ENZYME AND CHEMICAL ENCAPSULATION IN POLYMERIC MICROCAPSULES[a]

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Abstract

Polypyrrole microcapsules (prepared via the template method) were used for immobilization of both enzymatic and chemical catalytic systems. Enzymes immobilized include glucose oxidase, catalase, trypsin, subtilisin, and alcohol dehydrogenase. The chemical catalytic system investigated consisted of immobilized Pd nanoparticles for catalysis of hydrogen peroxide decomposition. Microcapsules loaded with glucose oxidase (GOD) were found to have higher enzymatic activity than GOD-loaded thin films, a competing encapsulation method. Trypsin was used to explore the possible leakage of small proteins form the capsules; no leakage was observed. Subtilisin was used to show that these microcapsules can be used in non-aqueous solvents. The effect of capsule wall thickness on the rate of enzymatic reaction was also explored.

Introduction

Enzyme immobilization has emerged as an important method for the development of biosensors and bioreactors.[1-20] Current methods of enzyme immobilization include adsorption or covalent attachment to a support,[2-4] microencapsulation,[5,6] and entrapment within a membrane/film[7,8,11-20] or gel.[9] An ideal immobilization method should employ mild chemical conditions, allow for large quantities of enzyme to be immobilized, provide a large surface area for enzyme-substrate contact within a small total volume, minimize barriers to mass transport of substrate and product, and provide a chemically and mechanically robust system.

We have recently developed a new approach for immobilization of enzymes. This approach entails immobilization of the desired enzyme in template-synthesized[21-26] polymeric tubules. While the template-approach allows for easy preparation of such tubular structures,[21-24,27-29] the tubules typically obtained are open on both ends and, therefore, not useful for enzyme immobilization. In this paper, we describe in detail[30] synthetic strategies to yield capped versions of such microtubules (that is, microcapsules). These microcapsules are prepared from the polymer polypyrrole and are arranged in a high-density array in which the individual capsules protrude from a surface like the bristles of a brush. We have developed procedures for filling these polypyrrole microcapsules with high concentrations of enzymes. The enzyme-loaded microcapsule arrays function as bioreactors in both aqueous solution and organic solvents.

Experimental Section

Materials. The monomer, pyrrole (Aldrich 99 %), was distilled prior to use. Purified water, obtained by passing house-distilled water through a Milli-Q (Millipore) water purification system, was used to make all solutions. $FeCl_3$ was used as received. Poretics microporous polycarbonate membrane filters (Poretics Corp) were used as the template membranes.[21-24] The membranes used for these studies had a pore diameter of 400 nm, a pore density of $1x10^8$ pores/cm^2, and were *ca.* 10 μm in thickness. Unless otherwise noted, the enzymes and substrate molecules were used as received, and enzyme solutions were prepared using a pH = 7.0 phosphate buffer (0.05 M).

Formation of the polymeric microcapsules. We have shown that when polypyrrole is synthesized from the monomer in the presence of the polycarbonate template membrane, the polymer preferentially nucleates and grows on the pore walls.[21,31] This leads to the formation of polypyrrole tubules within the pores of the membrane. However, these tubules are not capped and are, therefore, not useful for enzyme immobilization. The primary challenge in developing an immobilization method is developing methods for capping these tubules.[30] The capped tubules used in most of the studies described here were prepared using a method that combines both electrochemical and chemical synthesis of polypyrrole within the template membrane. We have also developed a purely chemical method for preparing the desired polypyrrole microcapsules . Both of these methods are

described here.

The Electrochemical/Chemical method. Unless otherwise noted, this is the procedure that we have used for most of the work reported here. A schematic of this strategy for preparing polypyrrole microcapsules is shown in Figure 1. Polypyrrole was synthesized using a two step procedure as follows: First, polypyrrole (PPy) "caps" are electrochemically[32] deposited at one face of the membrane. To accomplish this, one surface of the microporous polycarbonate template membrane is first sputtered with ca. 50 nm of gold (Figure 1a). The gold coats the surface of the membrane but does not block the pores. This thin gold layer serves as the electrode for the electrochemical polymerization of the PPy caps.

PPy was galvanostatically deposited at this Au surface film/electrode from a solution containing 0.2 M pyrrole and 1 M NaCl in 50:50 methanol:water mixture. The methanol was necessary to ensure proper wetting of the pores. A thick surface layer of PPy is deposited on the gold film electrode, and the polymer subsequently grows inwards into the pore. The PPy that grows into the pore will become the cap for the microcapsule. The thickness of this cap can be varied by varying the charge used during the electropolymerization. A charge of 0.21 C per cm^2 of membrane surface area was found to result in a cap of ca. 1 μm thickness within the pore (Figure 1b).

After electropolymerization of the PPy caps, PPy microtubules are synthesized chemically[21-24] within the pores of the membrane (Figure 1c). This is done by immersing the membrane in a solution containing 0.5 M

$FeCl_3$ as the oxidant and 0.2 M pyrrole as the monomer.[31] A polymerization time of 5 minutes was used. The PPy grows within the pores as well as on both surfaces of the membrane. The surface layers are removed by polishing the surfaces with 1 μm alumina powder using a Kim wipe wetted with methanol. The membrane is then sonicated and thoroughly rinsed in the pH = 7.0 buffer solution repeatedly to remove traces of monomer, oxidant and alumina powder. This two-step synthesis results in the formation of the desired capped tubules or microcapsules within the pores of the membrane (Figure 1c).

These PPy microcapsules are filled with enzyme by vacuum filtration of an enzyme solution through the capsule-containing membrane (Figure 1d). The microcapsules are first wetted with water by ultrasonicating the membrane in buffer solution. The membrane is then placed in a vacuum filtration apparatus with the cap-side down. A buffered solution of the enzyme is filtered through the open (upper) side of the membrane (Figure 1d). The water from the enzyme solution pervaporates through the polypyrrole cap.[33] The enzyme, being a large molecule, cannot permeate the PPy cap and is thus retained within the microcapsule.

Vacuum filtration is continued overnight until all the water has pervaporated through the PPy microcapsules. We have found that during this process, enzyme crystallizes on the upper surface of the membrane. This excess enzyme is wiped off by using a wet Kim wipe. Care must be taken while wiping excess enzyme from the surface because an excessively wet Kim wipe can wick the enzyme solution from the interior of the

microcapsule. Alternatively, a Kim wipe dipped in hexane can be used to remove the excess enzyme from the surface. Hexane is useful because the aqueous enzyme solution is not wicked out of the capsules. Torrseal epoxy is then applied to the face of the membrane that is not capped (Figure 1e). The epoxy is too viscous to go into the microcapsules. A glass rod is inserted into the epoxy so that the ensemble of microcapsule can be conveniently handled. Once the epoxy cures, the polycarbonate host membrane is dissolved using copious quantities of dichloromethane. This results in an ensemble of PPy microcapsules with enzyme contained within them (Figure 1f).

Finally, pieces of template membrane of 0.5 cm^2 surface area were used to prepare the microcapsule ensembles described here. This means that these ensembles contained $5x10^7$ enzyme-loaded microcapsules. Each microcapsule has an outside diameter of 400 nm, an inside diameter of 375 nm, a length of 10 μm (the thickness of the membrane), and a cap thickness of 1 μm.

Chemical method for microcapsule formation. A schematic of this method is shown in Figure 2. The polycarbonate membrane (Figure 2a) was immersed into 10 ml of a 0.2 M pyrrole solution and 10 ml of 0.5 M $FeCl_3$ were added.[31] Polymerization was allowed to proceed for 1 minute. During this period, a thin skin of PPy is deposited onto the walls of the pore (Figure 2b). The membrane was removed from this solution and rinsed in water. After drying, a piece of Scotch-brand tape was applied to one face of the

membrane. This tape prevents further polymerization of PPy on the face of the membrane covered by the tape.

The membrane/tape composite was then re-immersed into the polymerizing solution for an additional 30 minutes. During this time interval, the desired PPy microtubules are deposited within the pores of the membrane (Figure 2c). PPy was then removed from the surface of the membrane not covered by the tape using a wet Kim wipe. Polymerization was then repeated two more times by immersion of the membrane/tape composite into a polymerization solution that was 1M in $FeCl_3$ and 0.2 M in pyrrole. Polymerization was allowed to proceed for 5 minutes in the above two cases. During these polymerization periods, the PPy walls of the tubules thicken and ultimately caps of PPy are formed at the face of the membrane not covered with the tape (Figure 2d).

The surface PPy layer, on the cap-side is removed by polishing with alumina powder. The membrane was then cleaned by ultrasonication in water. Finally, the scotch- brand tape was detached from the membrane surface by repeated rinsing of the membrane in diethyl ether (Figure 2e). The ether dissolves the adhesive from the tape. At this stage, the PPy capsules can be loaded with enzyme as per Figure 1d.

Transmission electron microscopy (TEM). TEM images of the microcapsules prepared by the electrochemical/chemical method were obtained as follows: The microcapsule-containing polycarbonate membrane was placed on a copper TEM grid. The membrane was held in place by vacuum and was then washed with copious quantities of

dichloromethane. The dichloromethane dissolved the polycarbonate, leaving the PPy microcapsules on the TEM grid.

We have also attempted to image the encapsulated enzyme within the PPy microcapsule using TEM. The enzyme cannot be imaged directly because of insufficient contrast between the PPy and enzyme. However, a solution containing horse radish peroxidase (HRP) that is covalently attached to 20 nm colloidal gold particles is available from Sigma Chemical Co. This solution was used to load the microcapsules with Au-tagged HRP. Gold, being electron dense, can be easily observed within the PPy capsules using the TEM. The microcapsules used for these studies were prepared by the purely chemical method.

Monitoring Enzyme Activity. We have encapsulated five enzymes and one chemical catalytic system within microcapsules prepared via the electrochemical/chemical method described above. UV-visible spectroscopy was used to assay the activity of the encapsulated enzymes. The experimental set-up used is shown in Figure 3. The enzyme-loaded microcapsule array is inserted (using the glass rod) into a cuvette (3 mL) that contains the substrate for the enzyme and the enzyme assay chemistry (see below). The cuvette is present in the sample chamber of a Hitachi U-3501 UV-visible spectrometer. A magnetic stir bar is used to stir the solution in the cuvette. Prior to monitoring of enzyme activity, the enzyme-loaded microcapsule array was immersed into an aliquot of the buffer solution at 4° C. The solution was stirred for a period of 2 days. This allows any enzyme from defective tubes to leach out so that only enzymatic activity from

encapsulated enzyme is assayed.

Catalase. Catalase catalyses the decomposition of hydrogen peroxide to water and oxygen. The enzyme-mediated decay in hydrogen peroxide concentration can be conveniently monitored at 240 nm.[34] The catalase-loaded microcapsule array was introduced into a cuvette containing 3 mL of 35 mM H_2O_2 in the pH = 7.0 buffer. The decay in absorbance of hydrogen peroxide was monitored at 240 nm. A control experiment was done, wherein an ensemble of empty PPy capsules (i.e. with no catalase) was introduced into the cuvette solution. The response at 240 nm was again monitored as a function of time.

A simple test was performed to see if catalase leached out of the the PPy microcapsules during the enzymatic assay. The catalase-loaded microcapsules were introduced into the cuvette solution as described above, and the decay in absorbance at 240 nm was monitored. The catalase-loaded microcapsules were then removed from the cuvette solution and the monitoring of absorbance was continued. If no catalase leached from the capsules, no further decay in H_2O_2 concentration should be observed during the time interval that the capsules were removed from the solution. In contrast if catalase leached out, the H_2O_2 concentration should continue to decay even though the capsules are not present in the assay solution. This process of insertion and removal of the catalase-loaded microcapsules was repeated two times.

Glucose oxidase (GOD). The activity of encapsulated GOD was evaluated using the standard peroxidase-o-dianisidine assay method.[35]

GOD catalyzes the oxidation of glucose, in the presence of air, to yield gluconolactone and hydrogen peroxide. In the presence of the enzyme peroxidase, the H_2O_2 produced oxidizes the reduced form of dianisidine to the red oxidized form. The absorbance of the oxidized dianisidine is monitored at 500 nm.

In a typical experiment, the enzyme-loaded microcapsules were introduced into a cuvette solution containing glucose (92 mM), peroxidase (180 units) and o-dianisidine dihydrochloride (0.17 mM). The solution was stirred, and the absorbance of oxidized dianisidine was monitored as a function of time. This assay procedure was done at two levels of enzyme loading within the polypyrrole microcapsules - 0.0025 mg (0.45 units) and 0.1 mg (18 units). A control experiment with PPy capsules containing no GOD was also carried out.

The following experiment was conducted to determine the quantity of active GOD that is actually incorportated into the microcapsules. A solution containing a total of 0.1 mg of GOD was filtered through the microcapsule ensemble as described in Figure 1. The surface-deposited enzyme was then wiped away as before. The next step would typically be to apply torrseal to the membrane surface. However, for this experiment, the piece of membrane containing these GOD -loaded microcapsules was simply immersed into 2 ml of pH 7 buffer. This allowed the GOD that was loaded into the microcapsules to leach out of the capsules. The resulting GOD solution was assayed for its enzymatic activity.

Enzyme immobilization is important for the development of

biosensors.[11-20] A number of glucose biosensors have been made by entrapment of GOD within electrochemically grown PPy films. [11,12,14-20] In order to compare the microcapsule immobilization method to this more standard method, GOD-containing PPy were prepared electrochemically using the procedure described by Marchesiello and Genies.[12] GOD-loaded PPy films of two thicknesses (4.7 μm and 0.8 μm)[14] were prepared and evaluated. The immobilized enzyme activities of our microcapsules were compared with that of these electrochemically-grown films.

One of the important features of our microcapsule immobilization method is that the walls of the microcapsules are very thin. This should allow for facile diffusion of substrate and product molecules through the walls of these capsules. Unless otherwise noted, PPy capsules with wall thicknesses of ca. 25 nm-thick walls (corresponding to a polymerization time of 5 minutes) were used for these studies. However, in order to explore the effect of wall thickness on the kinetics of glucose oxidation, a set of experiments was done on microcapsules with walls that were ca. 90 nm thick (30 minute polymerization time).

Finally, while the majority of the experiments described here were done using microcapsules prepared via the electrochemical/chemical method, encapsulation in the chemically-prepared microcapsules was also studied. This was accomplished by loading the chemically-prepared capsules with 0.1 mg (18 units) of GOD and evaluating the enzymatic activity using the dianisidine/peroxidase method.

Trypsin. Trypsin is a relatively small enzyme (23,500 Daltons); it is, therefore, useful for investigating whether small proteins can be leached from the capsules through the capsule walls. Trypsin catalyses ester hydrolysis, and its catalytic activity was evaluated using Nα-benzoyl-L-arginine ethyl ester as the substrate molecule. The microcapsules were loaded with 1mg of trypsin in 0.5 mL of pH 7 buffer. The trypsin-loaded microcapsules were introduced into an aqueous solution of the substrate and the absorbance due to the product of the enzymatic reation (Na-benzoyl-L-arginine) was monitored at 253 nm. After the assay, these microcapsules were stored in pH 3 buffer at 4°C for a period of 45 days. After 45 days, the enzymatic activity of the microcapsules was re-evaluated. This was done to determine if the enzyme was retained within the microcapsule during the 45-day storage.

Alcohol Dehydrogenase (ADH). ADH catalyses alcohol oxidation using NAD^+ as a soluble cofactor. Because NAD^+ is a large molecule (660 Daltons), the ADH/NAD^+ system can be used to investigate whether molecules of this size can permeate through the PPy capsule wall. This was accomplished by loading the ADH (without NAD^+) into the capsules and then immersing the capsules into a solution containing ethanol (6.5%) and NAD^+ (1.4 mM). In order for the enzymatic reaction to occur, ethanol and NAD^+ must diffuse through the capsule wall and find the ADH within the capsule.

The enzymatic reaction was followed spectroscopically by monitoring the NADH absobance of the external solution at 340 nm.[34] Note, again, that after reaction with the enzyme the NADH must diffuse through the capsule

wall into the external solution. Finally, analogous experiments were conducted with an NAD^+ that was covalently-bound to a 40K Dalton dextran chain (Sigma Chemical Co). As before, the NAD^+/dextran was added only to the external solution. We anticipated that, in this case, no enzymatic reaction would be observed because the dextran chain would prevent the NAD^+ from permeating the capsule wall.

Subtilisin. Subtilisin catalyses transesterification reactions. Klibinov and coworkers[36] have carried out reactions catalysed by subtilisin in non-aqueous media. They have shown that the enzyme retains its activity even when it is not soluble in the solvent medium. Because PPy is insoluble in nearly all solvents, our microcapsules might be useful for such nonaqueous enzymatic processes. To explore this point, we investigated a transesterification reaction catalysed by subtilisin in acetone.

The subtilisin was loaded into the microcapsules from aqueous buffer solution. After evaporation of residual water, the capsules were immersed into an acetone solution of N-acetyl L-phenyl alanine ethyl ester (5 mM) and propanol (1 M). This transesterification reaction was monitored using a gas chromatography-mass spectrometry (GC-MS) method. An HP GC (5890 A) (with a DB-1 column) and an HP MS (5970) were used. GC conditions used were as follows: Detector temperature: 290°C, injector temperature: 275°C, oven temperature: 200°C, temperature ramp: 10°C/min for 5 minutes. The progress of the reaction was monitored as the ratio of area under the product (N-acetyl L-phenyl alanine propyl ester) peak to the area under the reactant (N-acetyl L-phenyl alanine ethyl ester) peak.

Palladium nanoparticles. In addition to biocatalysts, we have shown that these microcapsules can be used to encapsulate a chemical catalytic system. Palladium nanoparticles were prepared by mixing equal volumes of a solution of Palladium(II) chloride (0.01M) and a solution of $SnCl_2$ (0.025 M) in 0.1 M HCl.[37] The resulting Pd nanoparticles were then filtered through the PPy microcapsules (as per Figure 1d). The encapsulated Pd nanoparticles were used to catalyse the decomposition of hydrogen peroxide, using the methods described for the enzyme catalase.[34]

Results and Discussion

Electron Microscopy. Figure 4a shows a scanning electron micrograph (SEM) of typically PPy microcapsules prepared via the electrochemical/chemical methods. This SEM shows that the microcapsules are capped and that the capsules protrude from the torr seal surface like the bristles of a brush. The exposed PPy surface area in such a microcapsule array is 12 cm^2 of capsule surface area per cm^2 of substrate epoxy area. This figure could be increased by using membranes with higher porosity (the porosity of this membrane is only 12 %) or by using thicker membranes (and thus making longer micro capsules).

The wall thickness in these microcapsules can be controlled by varying the polymerization time. Unless otherwise noted, a polymerization time of 5 mins was used for these studies. This yields a wall thickness of ca. 25 nm (see Figure 4b). These thin walls ensure that diffusion of substrate and product molecules through the capsule is facile. In spite of these thin walls, these capsules have extraordinary mechanical strength.[38] Shorter

polymerization times yield microcapsules with poor mechanical strength, and broken or flattened capsules are observed in the TEM after dissolution of the membrane. Longer polymerization times cause diffusional mass transport through the capsules to be slower (see below).

While the electrochemical/chemical route for preparing microcapsules allows for the greatest amount of control over the capsule growth process, this method requires two separate synthetic steps, and the electrochemical step is more difficult than the chemical step. We have, therefore, investigated purely chemical routes for preparing such microstructures. We have found that if the membrane is immersed in the polymerization solution for long times (2 hours) the tubules formed ultimate cap on both ends to yield bottle-like microstructures (see Figure 4c). While these are interesting microstructures, the caps on both ends make it difficult to load these structures with enzyme, unless of course, the enzyme is co- dissolved in the polymerization solution. This is undesirable because the pH of this solution is quite low (pH ca. 1).

This led us to idea of masking one surface of the membrane with scotch tape. By using this approach, caps cannot form on the masked surface and open microcapsules are obtained. These microcapsules can then be filled with enzyme via the simple filtration method described in Figure 1. Figure 4d shows a TEM of microcapsules prepared via this chemical approach. These capsules had been loaded with a low concentration solution of horse radish peroxidase that had been tagged with

a 20 nm Au nanoparticle. The Au nanoparticles are clearly evident in this TEM.

Catalase. When the catalase-loaded microcapsules were immersed into the cuvette solution containing hydrogen peroxide (Figure 5A, time a), a rapid decay in H_2O_2 absorbance was observed due to the catalytic decomposition of the H_2O_2. At time b, the capsule array was removed from the cuvette solution. The decay in H_2O_2 concetration slows to that corresponding to the natural (i.e. non-catalyzed) rate. This process was repeated two more times, and similar responses were obtained (Figure 5A). This indicates that the catalase does not leach out of the microcapsules . If catalase leached from the microcapsules, then the higher (catalytic) rate of decay would be observed after the capsules were removed, from the cuvette. Finally, as a control, empty PPy capsules were introduced into the cuvette solution. As expected, little change in absorbance (corresponding to the natural decay of H_2O_2) is observed (Figure 5 B).

Glucose oxidase. The standard o-dianisidine-peroxidase assay procedure was used to determine the catalytic activity of GOD within the microcapsules. Curves a and b in Figure 6 compare catalytic activities of two different loadings of glucose oxidase within the microcapsules: 0.1 mg (18 units) for curve a and 0.0025mg (0.45 units) for curve b. As would be expected, the capsules with the higher GOD content show higher enzymatic activity. This ability to control the amount of enzyme immobilized within the microcapsule is an important feature of this immobilization method. Curve e in Figure 6 is the response obtained when no enzyme is present within the

microcapsule. As would be expected, no enzymatic activity is observed.

An assay of the quantity of GOD loaded within the capsules used to obtain the data in curve a showed that an amount equivalent to 625 mg of GOD per ml of capsule volume was present in each capsule. It can be shown from the specific volume of glucose oxidase,[39] that this amount corresponds to 47% of the available volume within the microcapsule. This shows that large quantities of enzyme can be loaded with these microcapsules.

A number of proposed glucose biosensors have been prepared by physically entrapping GOD within polypyrrole films.[11,12,14-20] Curves c and d in Figure 6 show the enzymatic activities of two such PPy films.[11,12] The activities for these films were nearly the same because only a thin layer (ca. 0.3 μm thick) at the outer surface of the polypyrrole film is enzymatically active.[13,15] A comparison of the slopes of curves c and d (competing immobilization method) with the slope of curve a clearly shows that higher enzymatic activity can be achieved with the microcapsule-immobilization method. Thus, these microcapsule ensembles may show promise for the development of new types of enzymatic biosensors.

The effect of the PPy wall thickness on the kinetics of glucose oxidation was also investigated. In all the experiments described so far, capsules with a wall thickness of 25 nm were employed. These thin walls minimize diffusional mass- transport barriers. To explore this point more fully, capsules with 90 nm-thick walls were also investigated. Figure 7

shows plots of rate of the enzymatic reaction vs. concentration of glucose for capsules with 25 nm and 90 nm-thick walls. Note the dramatically slower rate for the capsules with the thicker walls. Assays of the quantity of enzyme loaded indicated that both the 90 nm-thick and 25 nm-thick capsules contained the same amount of GOD. Hence, the lower rate for the thicker-walled capsules is due to slower mass transfer. Finally, in both cases, enzymatic rate ultimately levels at high substrate concentration indicating that typical Michaelis-Menten behaviour is observed for these capsules.

Microcapsules prepared by the purely chemical route were also used to encapsulate GOD. That the GOD in these capsules is enzymatically active is shown by the data in Figure 8. We currently prefer the electrochemical/chemical method for preparing such microcapsules. This is because it is easier to control the wall thickness in the chemical step after the caps have been formed electrochemically. However, it seems possible that with careful attention to polymerization time and conditions, procedures could be devloped that yield optimal microstructures from the simpler purely chemical method.

Trypsin. The encapsulated trypsin was used to catalyse the hydrolysis of Nα-benzoyl-L-arginine ethyl ester. An increase in absorbance was observed at 253 nm corresponding to the formation of the product Nα-benzoyl-L-arginine (Figure 9, curve a). When the catalytic activity was re-evaluated after 45 days, no loss of enzymatic activity was observed within experimental error (Figure 9, curve b). These results indicate that trypsin is

not leached out of the capsules. Again, as trypsin is one of the smallest of enzymes, this means that nearly any enzyme can be permanently immobilized within these microcapsules.

Alcohol Dehydrogenase. ADH was loaded into the microcapsules and its catalytic activity was measured by monitoring the NADH produced (at 340 nm) in the external solution. An increase in absorbance at 340 nm was observed corresponding to the formation of NADH (Figure 10 curve a). The only way that NADH could have been detected in the external solution is by alcohol and NAD^+ diffusing through the walls of the capsule and reacting with the encapsulated ADH. These experiments suggest that even large molecules such as NAD^+ and NADH can permeate through the walls of the capsules. Because substrate molecules for most enzymatic processes are smaller than this, these experiments show, again, that these capsules can be used with nearly any enzymatic system. It is possible for large molecules to permeate the capsule wall because polypyrrole is nanoporous[40] and because the walls are thin. In another experiment, dextran-linked NAD^+ was used in the external solution. No enzymatic activity was observed (Figure 10 curve b). This is because the large dextran chain (Mol.Wt=40,000) cannot diffuse through the walls of the PPy microcapsule.

Subtilisin. The transesterification reaction catalysed by the encapsulated subtilisin was monitored using GC-MS. Figure 11 shows typically GC MS data and compares the chromatogram at time = 0 and at 67 hours into the course of the enzymatic reaction. The appearance of the product ester is clearly seen. The progress of the reaction was monitored by ratioing the area of the product

ester peak to the area of the reactant peak. A plot of progress of the reaction versus time is shown in Figure 12 for both subtilisin-loaded and empty microcapsules. No reaction is observed for the empty capsules. Finally, this reaction occurs at a much slower rate than any of the other enzymatic processes investigated. This is because the the transesterification reaction is slow in the absence of water. [36] The upward curvature in the progess vs. time plot for the subtilisin-loaded capsules is undoubtedly due to the incorporation of water into the acetone from water vapor in the air.

Palladium nanoparticles. That the Pd nanoparticles incorporated into the microcapsules are catalytically active is proven by the data in Figure 13. A rapid decay in H_2O_2 concentration is observed upon immersion of the Pd-loaded microcapsules. The decay goes back to the slow background rate when the microcapsules are removed from the solution.

Conclusion.

Polymeric microcapsules have been prepared and evaluated as encapsulating agents for bio and chemical catalysts. The polymer used to pepare these capsules - polypyrrole - has several important attributes. First, it is known to be nanoporous and this allows for facile diffusion of substrate and product molecules through the capsule walls. Our work has shown that the molecular weight cut-off for the nanopores in polypyrrole is between 660 Daltons (NAD which does permeate the capsule wall) and 23,000 Daltons (trypsin which does not permeate the capsule wall). This molecule weight cutoff makes this polymer ideal for enzyme immobilization. The second important feature of this polymer

is its good mechanical properties. We have shown that tubular PPy microstructures with extremely thin walls have remarkable mechanical strength.[24] This ability to make encapsulants with extremely thin walls, again, minimizes mass transport barriers in these encapsulation systems.

The encapsulation method we have developed offers a number of important advantages. First, it is very general. Any enzyme or combination of enzymes can be loaded into these microstructures. Second, the enzyme-loading process is chemcially benign. The enzyme nevers sees harsh chemical environments. Finally, this encapsulation method provides for a large surface area for enzyme/substrate contact. In the current system, there are 12 cm^2 of encapsulant surface area per cm^2 of substrate area. This number could, in fact, be significantly higher. For example, an Anopore microporous alumina membrane has a porosity of ca. 55%, a pore diameter of 200 nm, and a thicknesses of 55 μm. If analogous microcapsules could be prepared in these membranes, an encapsulation system that offers 192 cm^2 of surface area per cm^2 of substrate area would be obtained.

We are currently exploring various possible applications for these microcapsule ensembles. We are particularly interested in applications in biosensors.

Acknowledgements. This work was supported by the Office of Naval Research.

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Figure captions.

Figure 1. Schematic diagram of methods used to synthesize and enzyme-load the microcapsule ensemble using the electrochemical / chemical route.

Figure 2. Schematic diagram of methods used to prepare microcapsule ensemble using the purely chemical route.

Figure 3. Experimental set-up used for UV-visible spectroscopy.

Figure 4A. Scanning electron micrograph of a typical microcapsule ensemble showing PPy microcapsules extending from the torr seal surface.

Figure 4B. Transmission electron micrograph of microcapsules that had not been attached to the epoxy surface. Scale bar in upper left corner is 1.0 μm.

Figure 4C. Transmission electron micrograph of free PPy microcapsules prepared using the chemical method. Scale bar in the upper left corner is 1.0 μm.

Figure 4D. Transmission electron micrograph of microcapsules that contain horseradish peroxidase that has been labeled with 20 nm-diameter gold particles.

Figure 5A. Absorbance due to H_2O_2 upon insertion (points a, a', a") and removal (points b and b') of a catalase-loaded microcapsule ensemble into a solution that was 35 mM in H_2O_2, pH = 7.0 phosphate buffer.

Figure 5B. Absorbance at 240nm after immersion of empty PPy microcapsules into a solution 30mM in H_2O_2.

Figure 6. (A) Absorbance for the oxidized form of o-dianisidine after immersion of glucose oxidase-loaded and empty microcapsule

ensembles into a glucose oxidase assay solution (see text for details).

Figure 7. Plot of rate vs. concentration of glucose for two different microcapsule wall thicknesses.

Figure 8. Absorbance for the oxidized form of o-dianisidine after immersion of chemically-prepared glucose-oxidase microcapsules into an assay solution.

Figure 9. Absorbance for N-α-benzoyl L-arginine when trypsin loaded capsules were immersed into the assay solution : Solid line is before and dashed line is after 45 day storage at 4°C.

Figure 10. Assay of alcohol dehydrogenase activity. Dashed line is the absorbance due to NADH upon insertion of alcohol dehydrogenase loaded microcapsules. Solid line is the analogous experiment done with dextran-linked NAD.

Figure 11. Gas chromatograms from the subtilisin-catalysed transesterification reaction mixture as a function of time.

Figure 12. Progress^{x} of transesterification reaction after immersion of enzyme-loaded and empty microcapsules into reaction mixture. Progress^{x} of reaction was monitored by ratioing the area under the product ester peak to the area under the reactant ester peak.

Figure 13. Absorbance at 240nm upon insertion (a), and removal (b) of Palladium-loaded microcapsules.

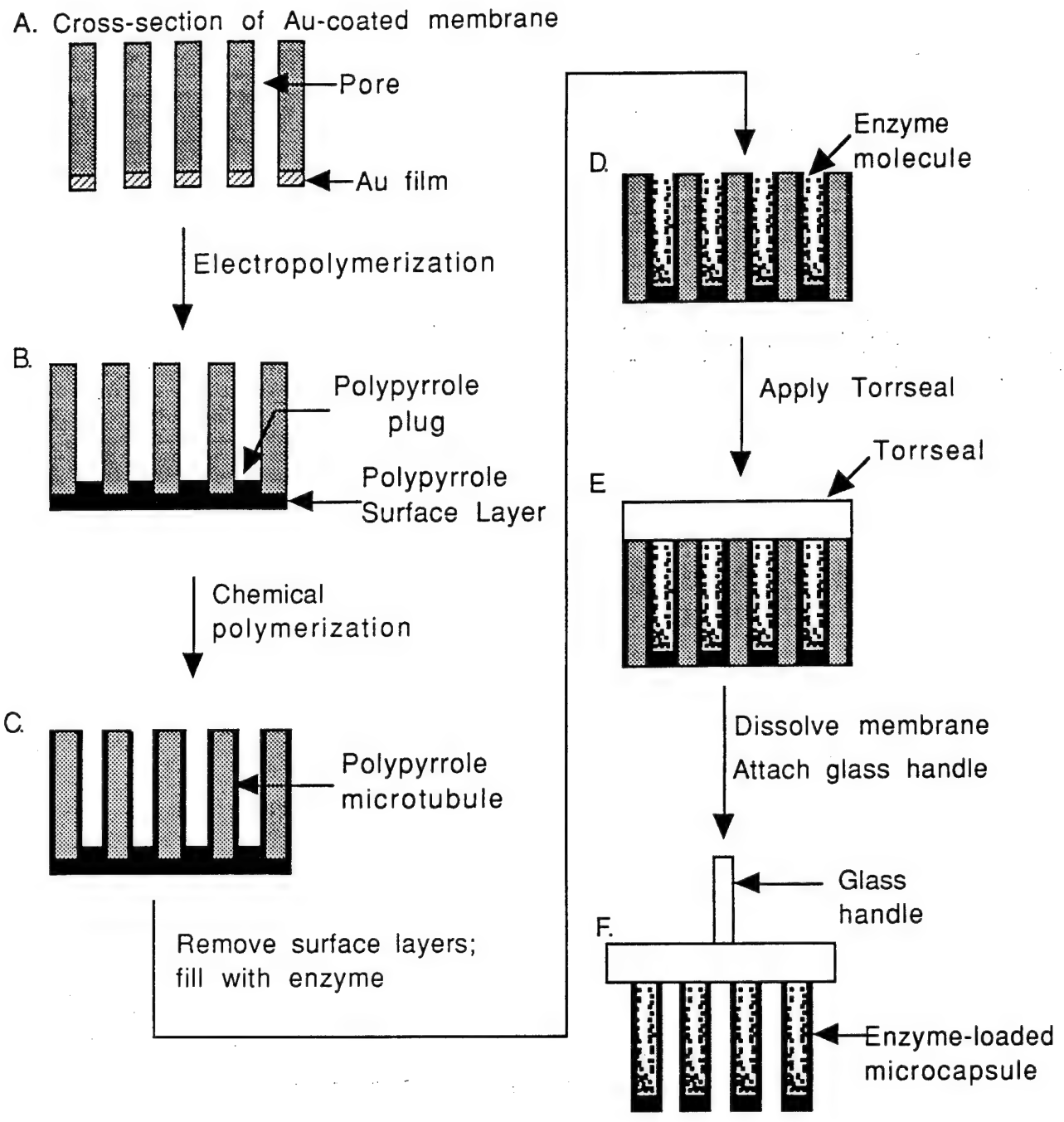

A. Cross-section of Au-coated membrane
Pore
Au film
Electropolymerization
B.
Polypyrrole plug
Polypyrrole Surface Layer
Chemical polymerization
C.
Polypyrrole microtubule
Remove surface layers; fill with enzyme
D.
Enzyme molecule
Apply Torrseal
E
Torrseal
Dissolve membrane
Attach glass handle
F.
Glass handle
Enzyme-loaded microcapsule

Fig 1

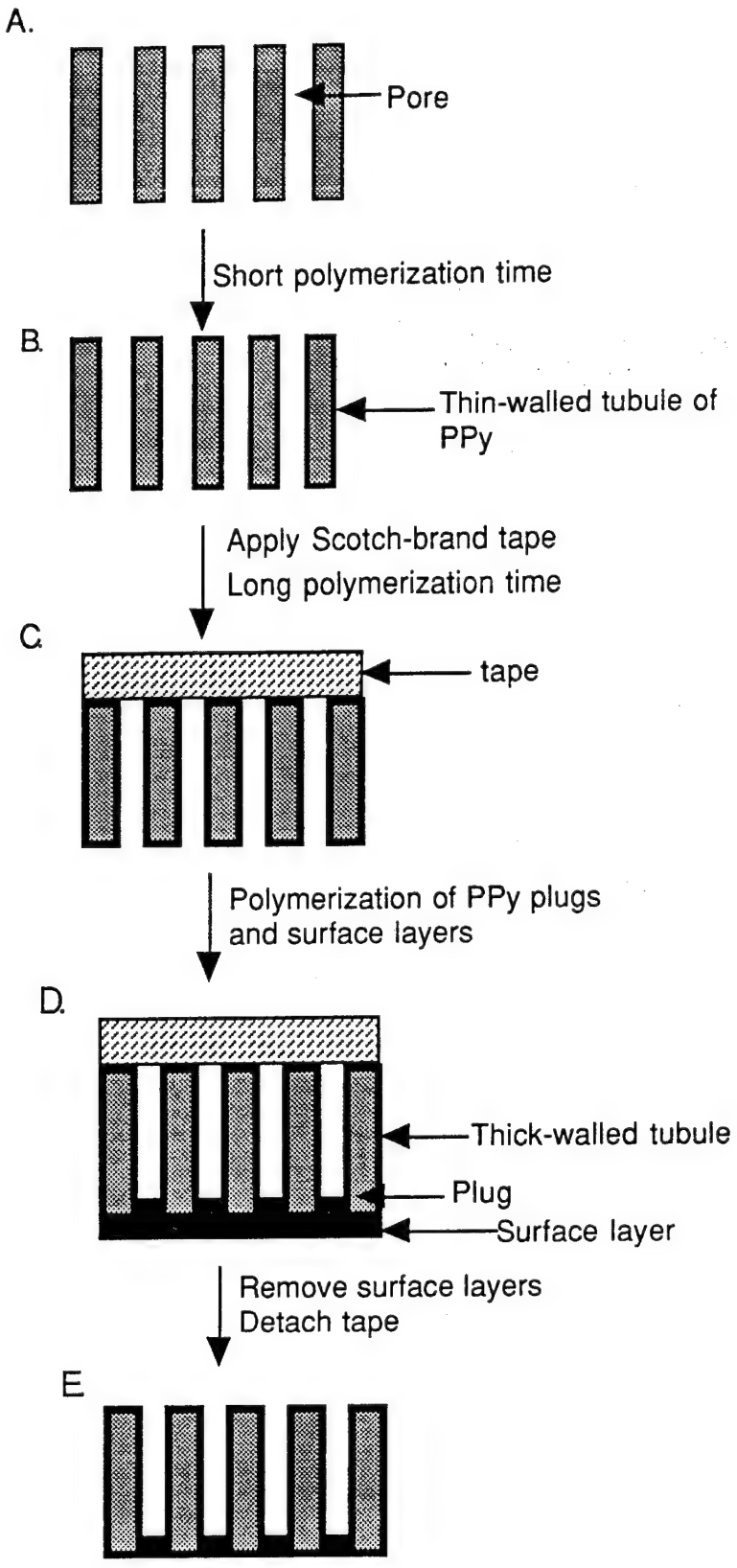
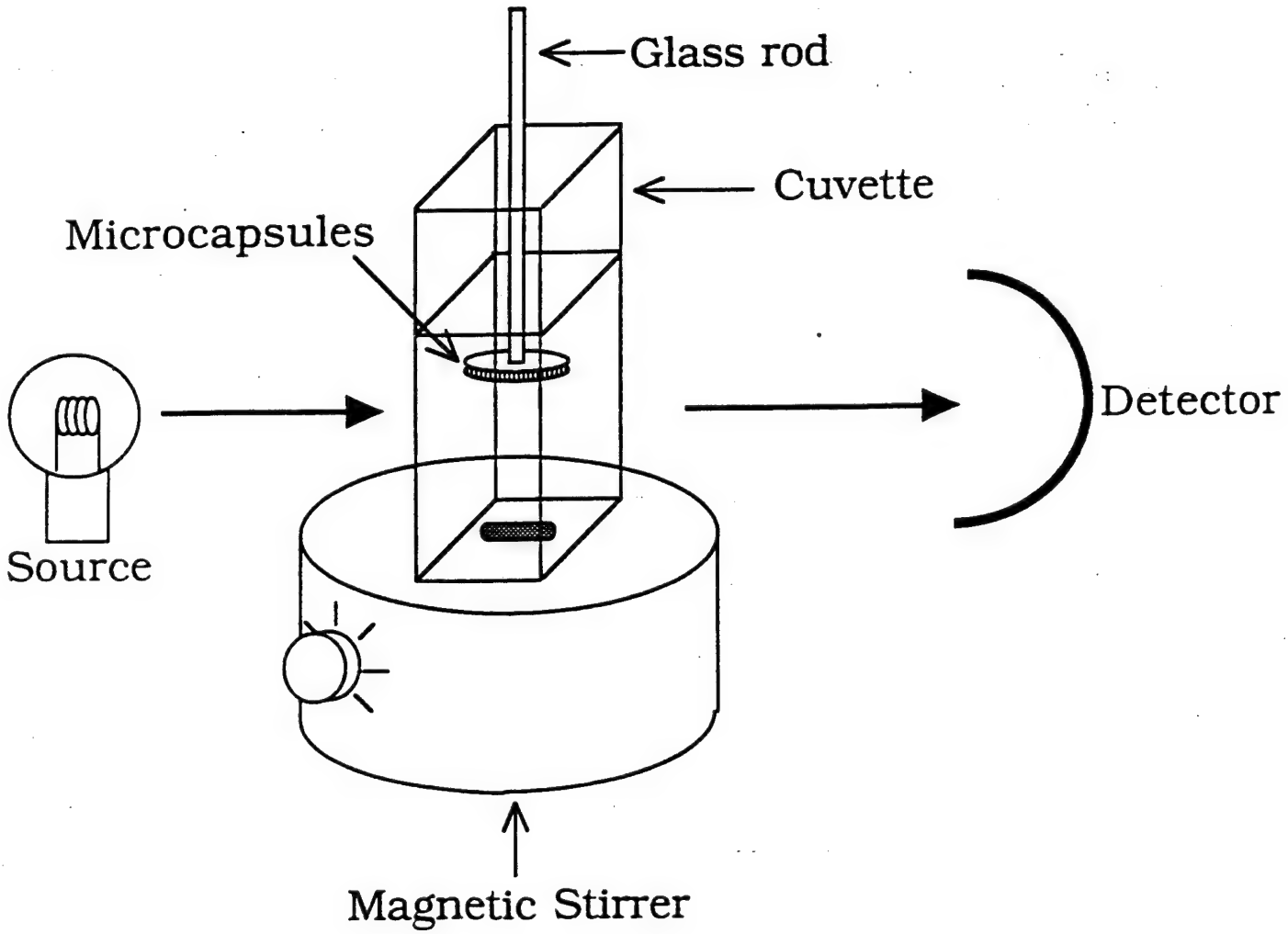

A.
Pore
Short polymerization time
B.
Thin-walled tubule of PPy
Apply Scotch-brand tape
Long polymerization time
C.
tape
Polymerization of PPy plugs and surface layers
D.
Thick-walled tubule
Plug
Surface layer
Remove surface layers
Detach tape
E.

Fig 2


Glass rod
Cuvette
Microcapsules
Detector
Source
Magnetic Stirrer

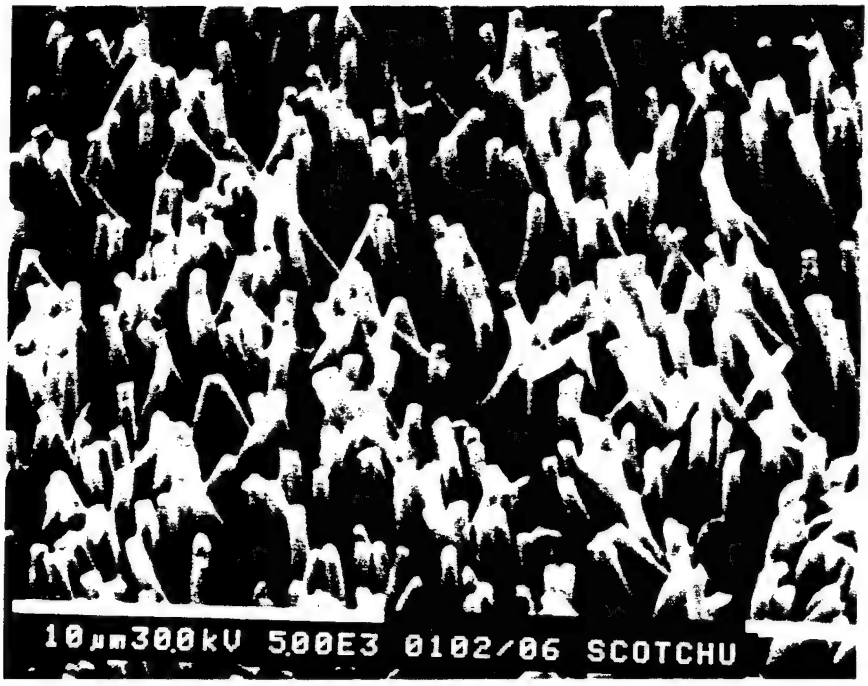


Fig 4

Fig 4b

Fig 4 C

Fe a.d

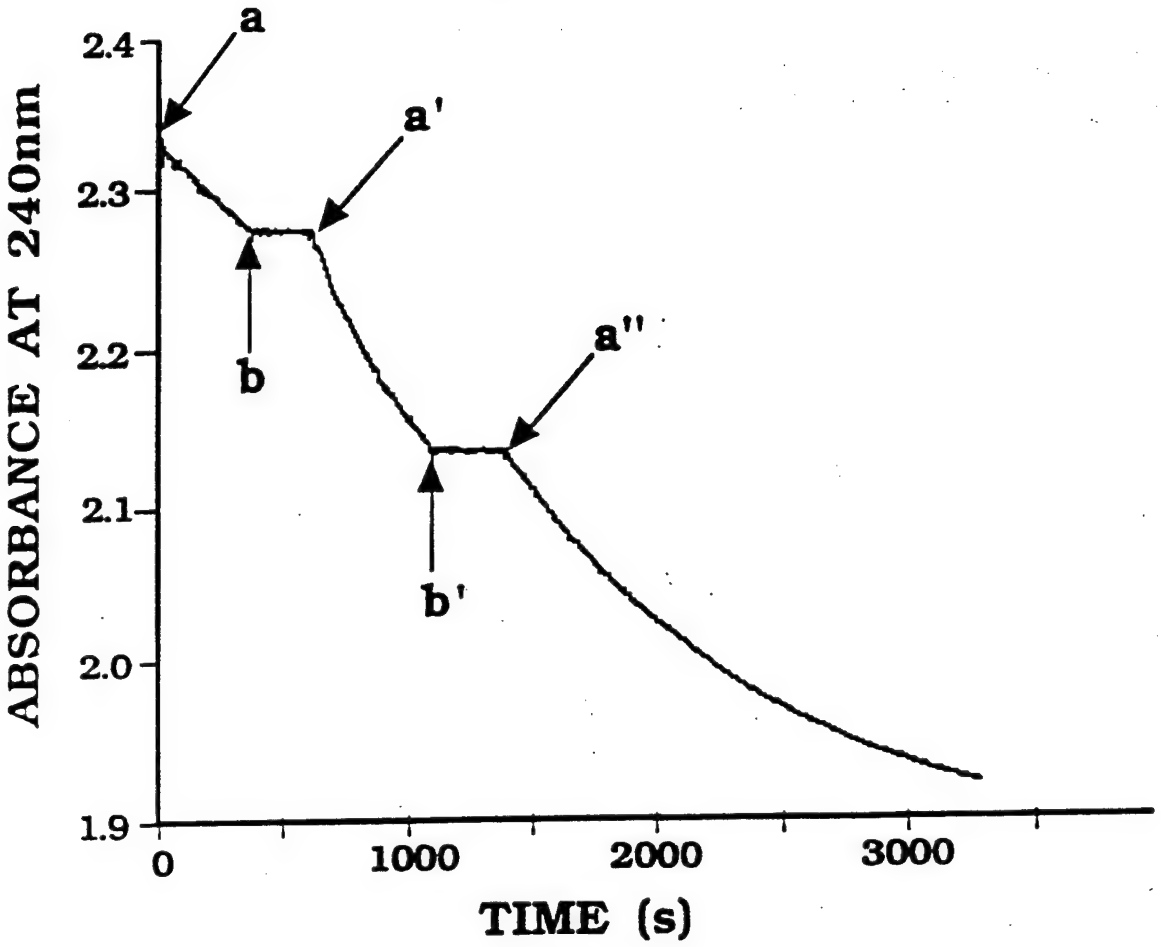

a
a'
a''
b
b'
2.4
2.3
2.2
2.1
2.0
1.9
ABSORBANCE AT 240nm
0
1000
2000
3000
TIME (s)

Fig 5A

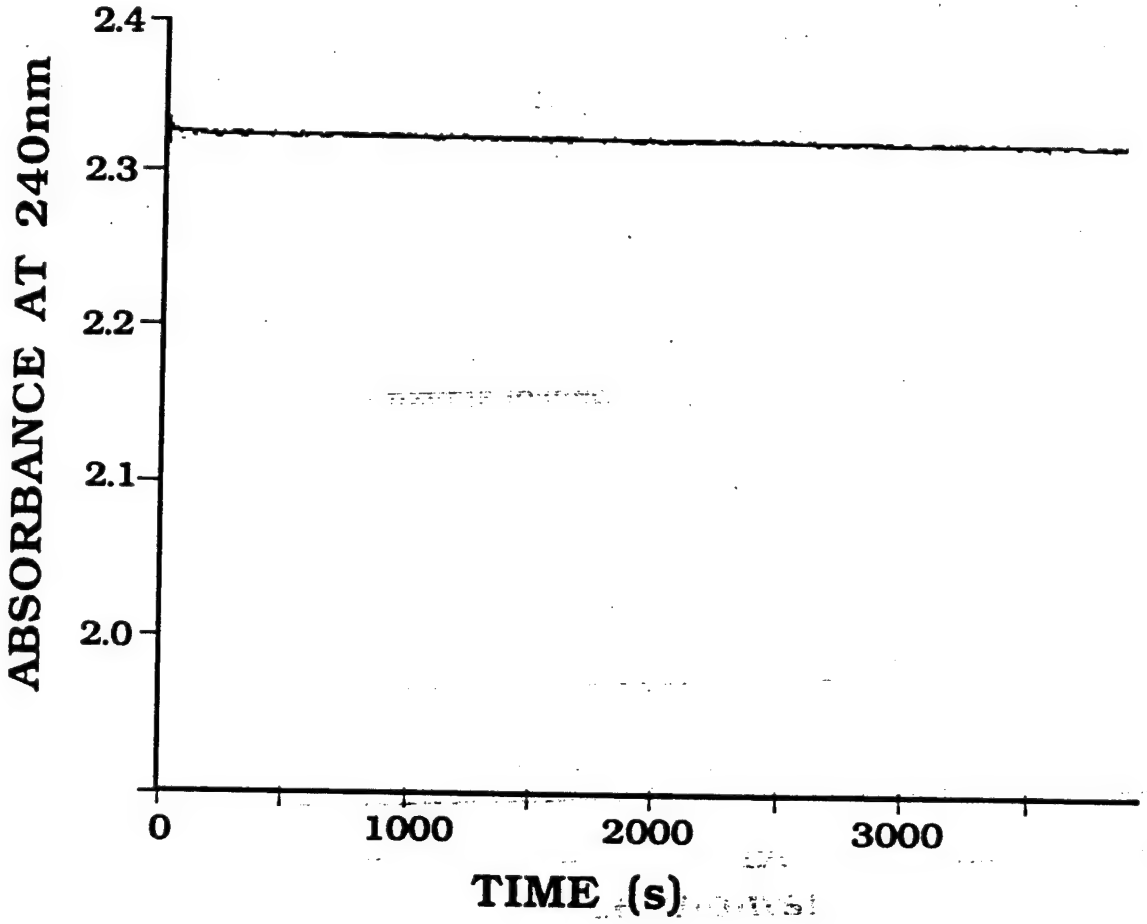
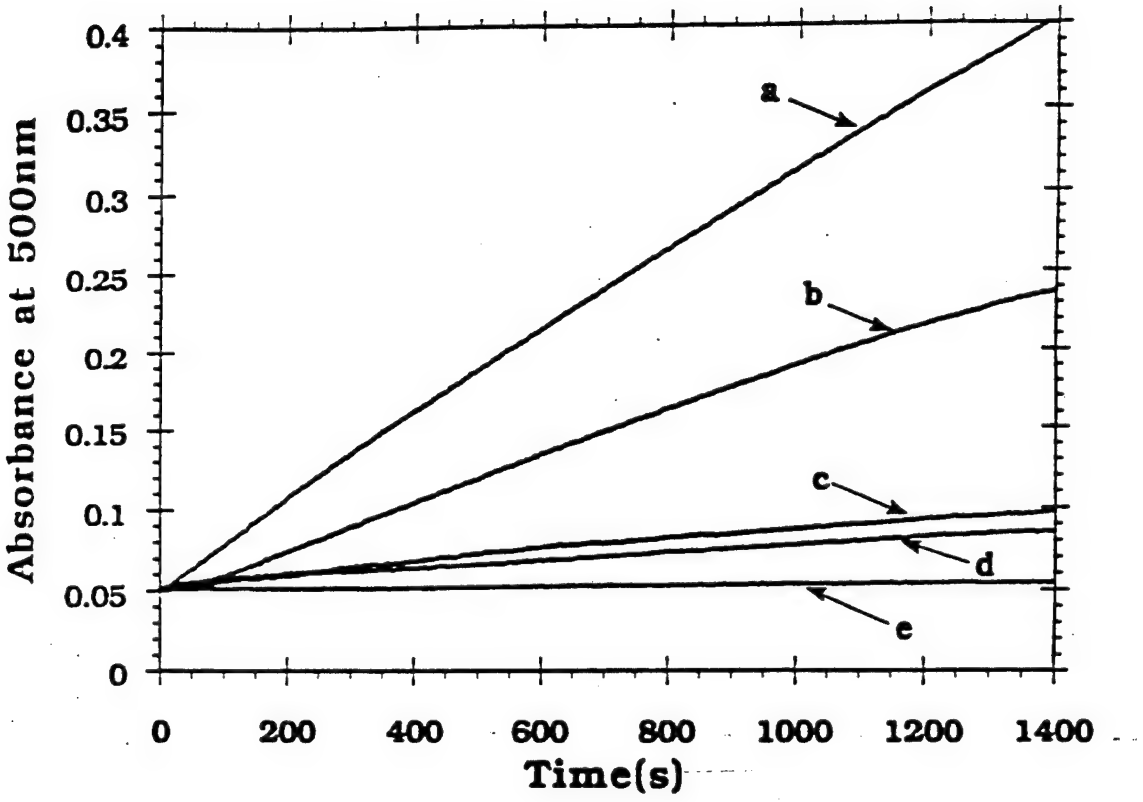


Fig 5E


Absorbance at 500nm
0.4
0.35
0.3
0.25
0.2
0.15
0.1
0.05
0
0
200
400
600
800
1000
1200
1400
Time(s)
a
b
c
d
e

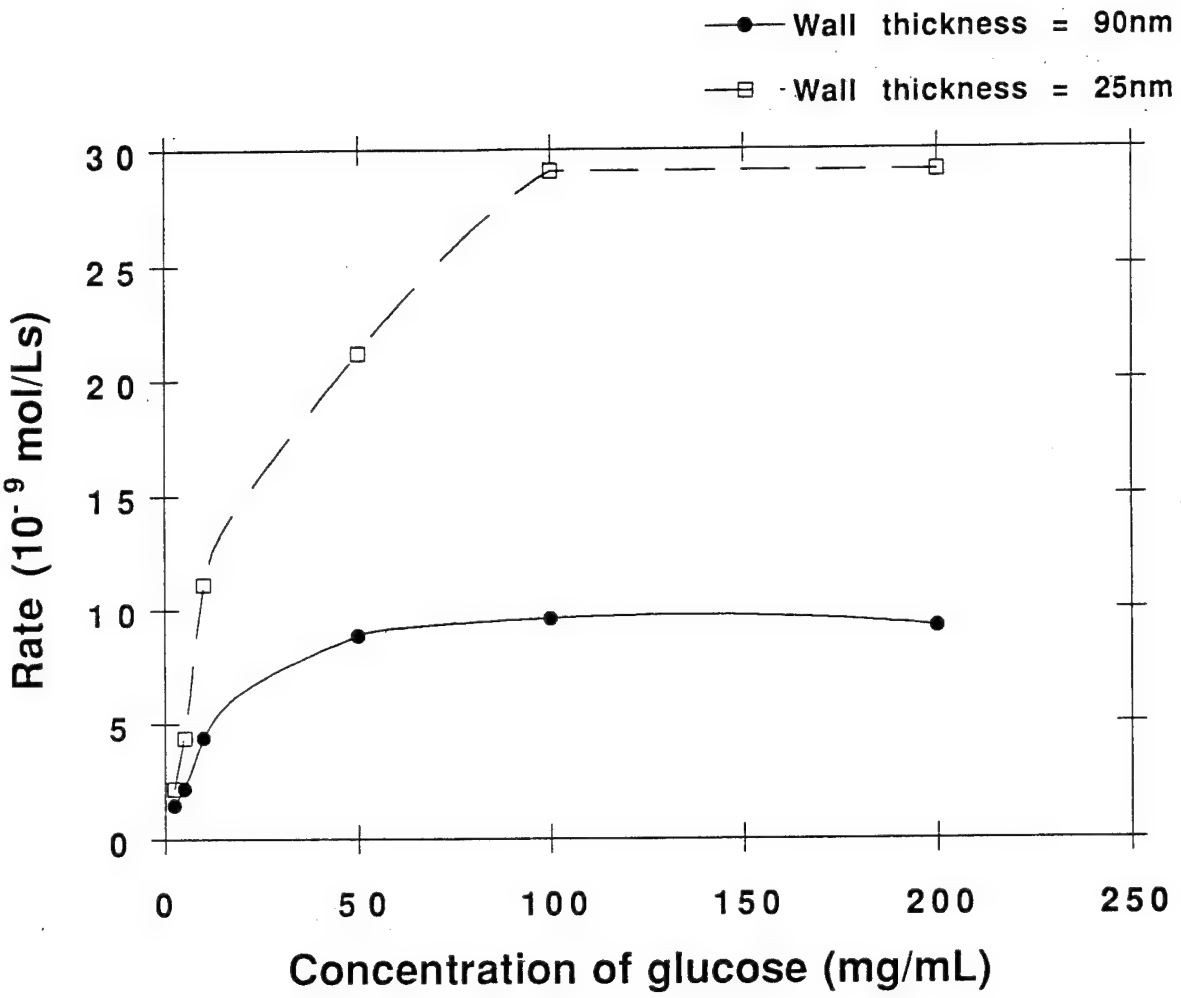

Wall thickness = 90nm
Wall thickness = 25nm
Rate (10^{-9} mol/Ls)
30
25
20
15
10
5
0
0
50
100
150
200
250
Concentration of glucose (mg/mL)

fig7

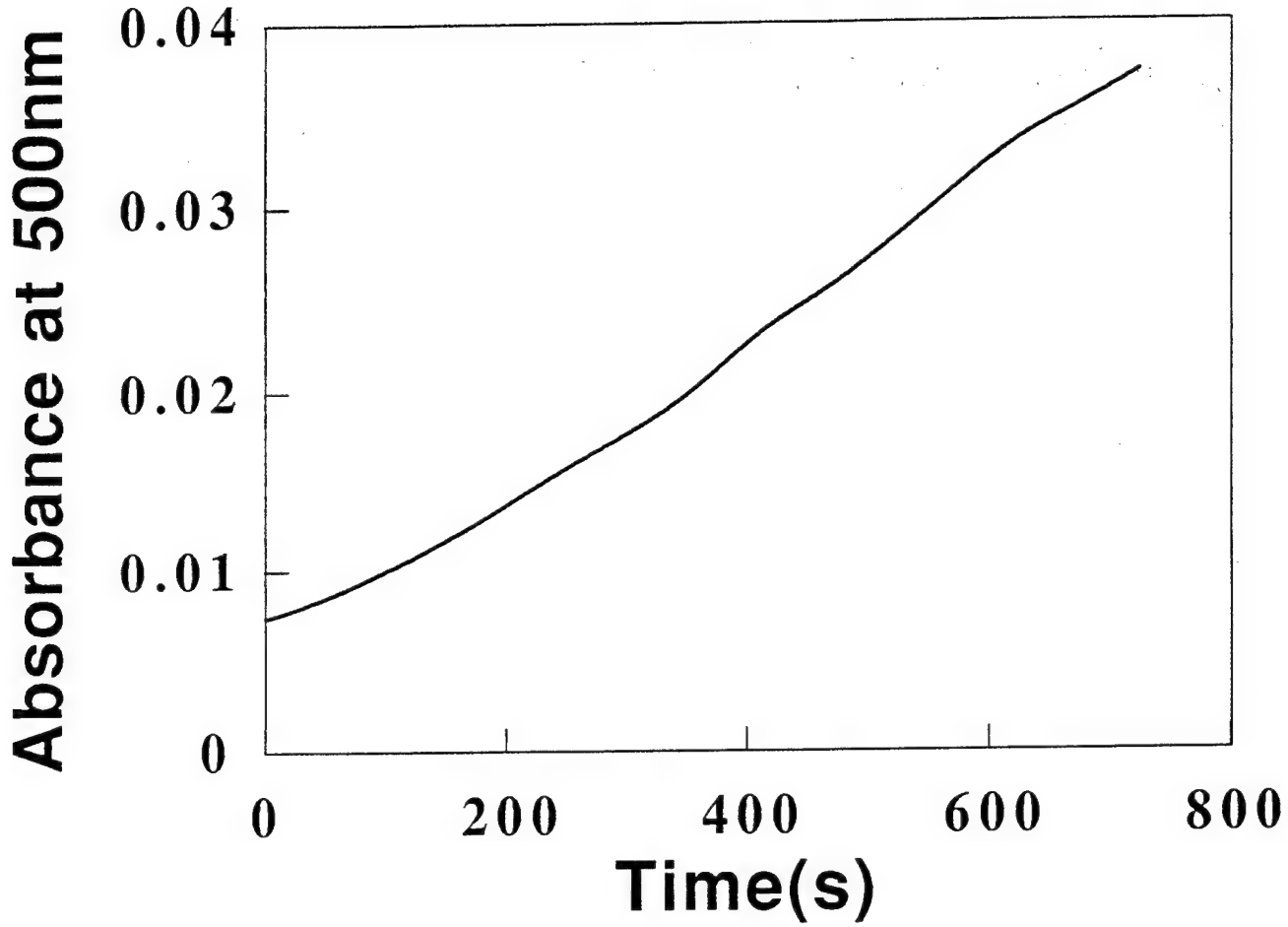
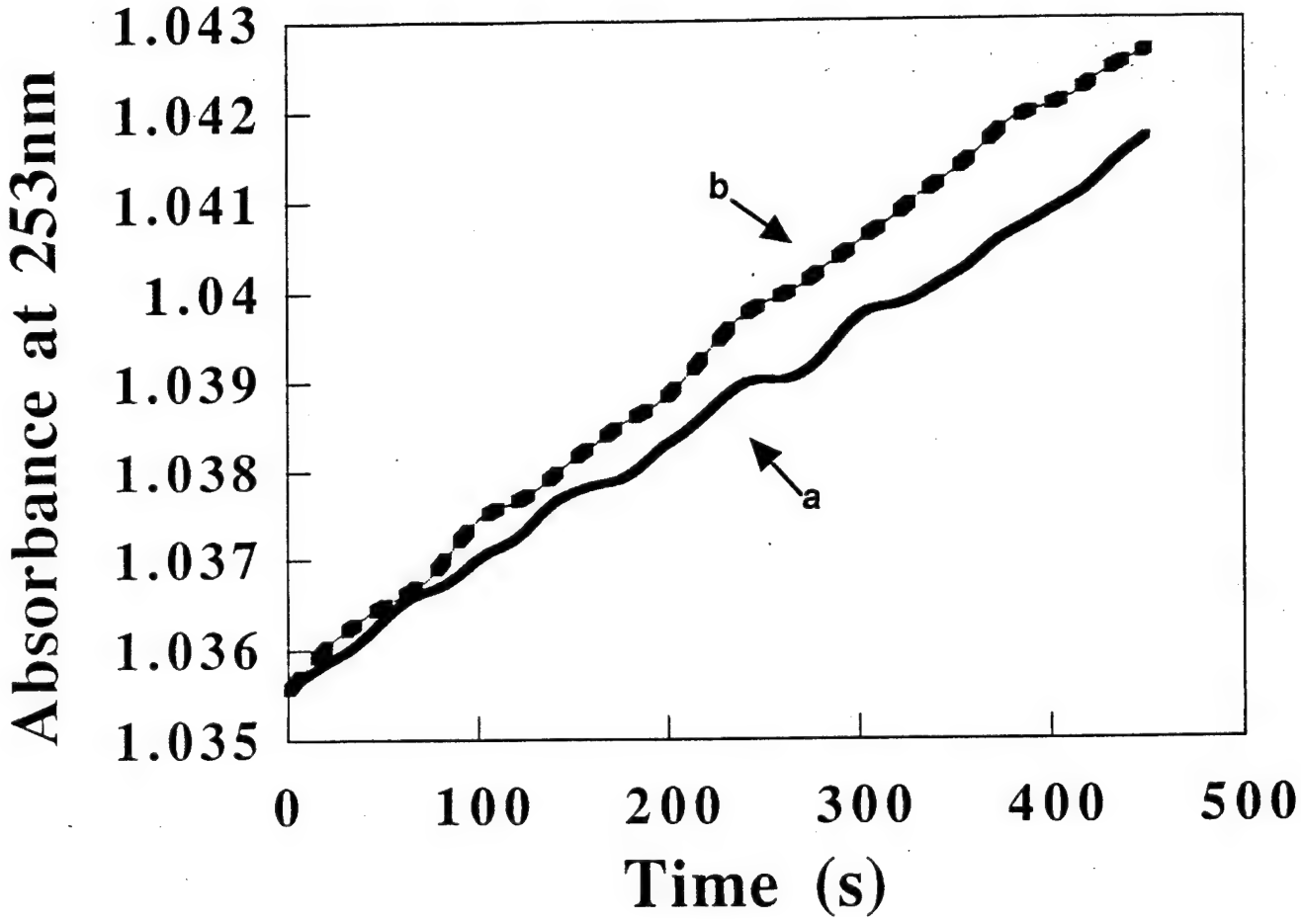


Fig 8


Absorbance at 253nm
1.043
1.042
1.041
1.04
1.039
1.038
1.037
1.036
1.035
0
100
200
300
400
500
Time (s)
b
a

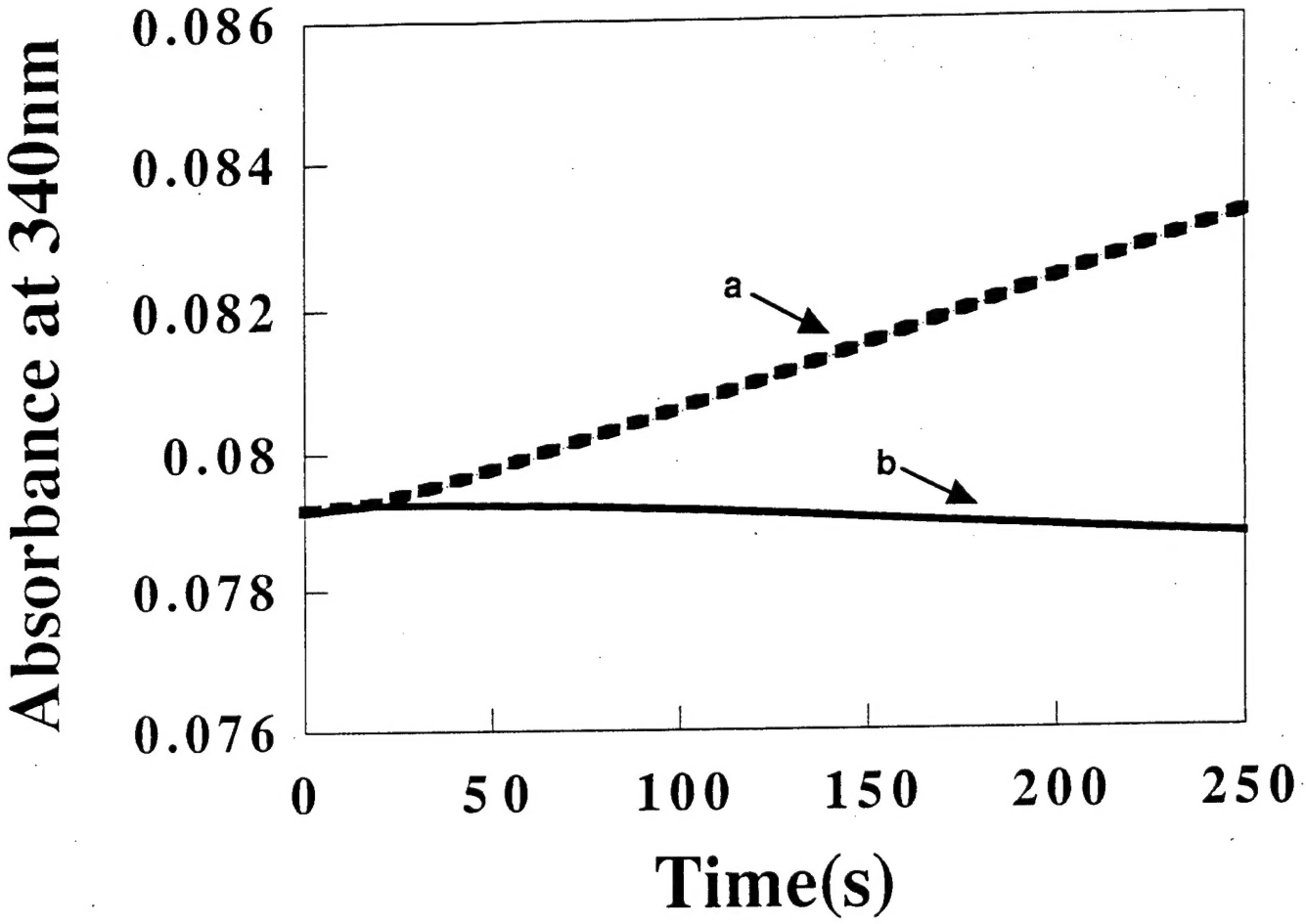
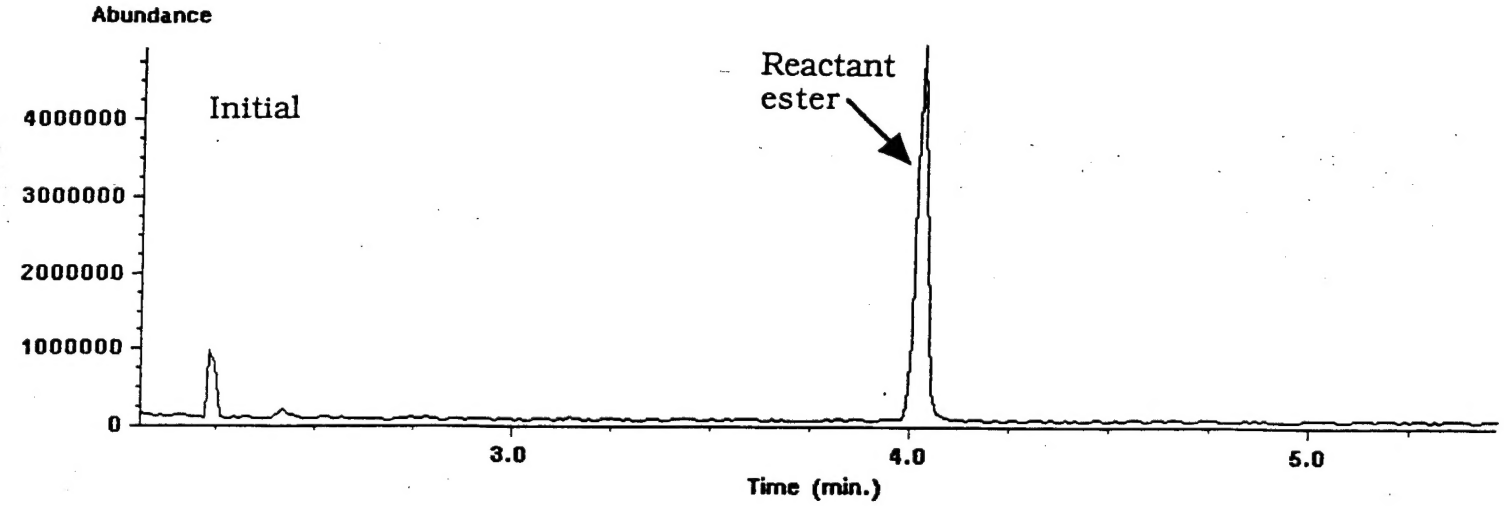
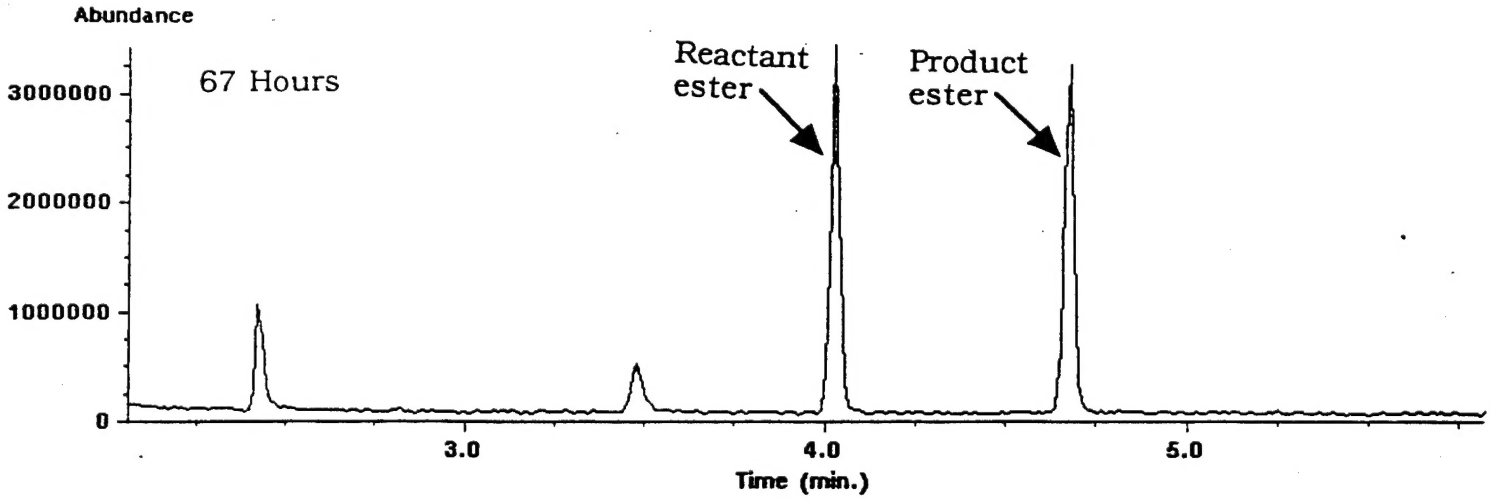
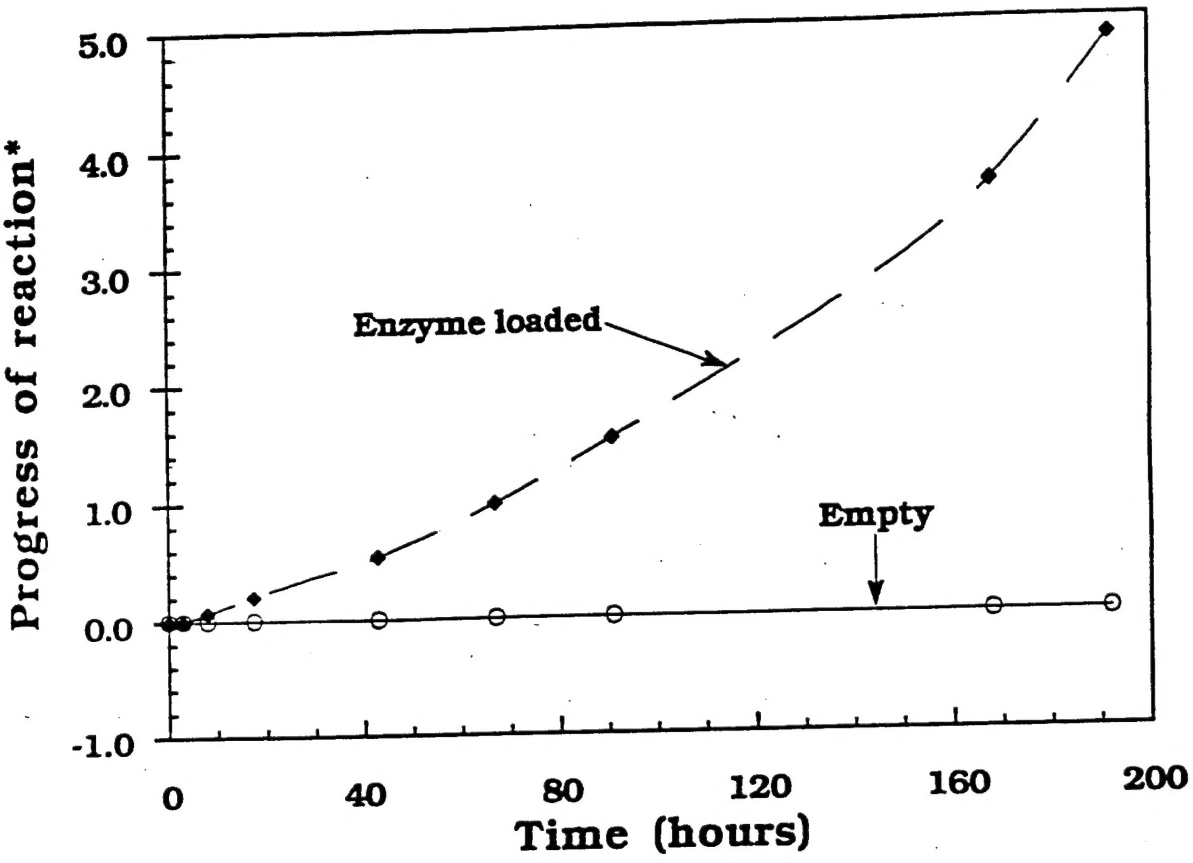

Absorbance at 340nm
0.086
0.084
0.082
0.08
0.078
0.076
0
50
100
150
200
250
Time(s)
a
b

Fig10


Abundance
Initial
Reactant ester
4000000
3000000
2000000
1000000
0
3.0
4.0
5.0
Time (min.)


Abundance
67 Hours
Reactant ester
Product ester
3000000
2000000
1000000
0
3.0
4.0
5.0
Time (min.)


5.0
4.0
3.0
2.0
1.0
0.0
-1.0
Progress of reaction*
Enzyme loaded
Empty
0
40
80
120
160
200
Time (hours)

F&B

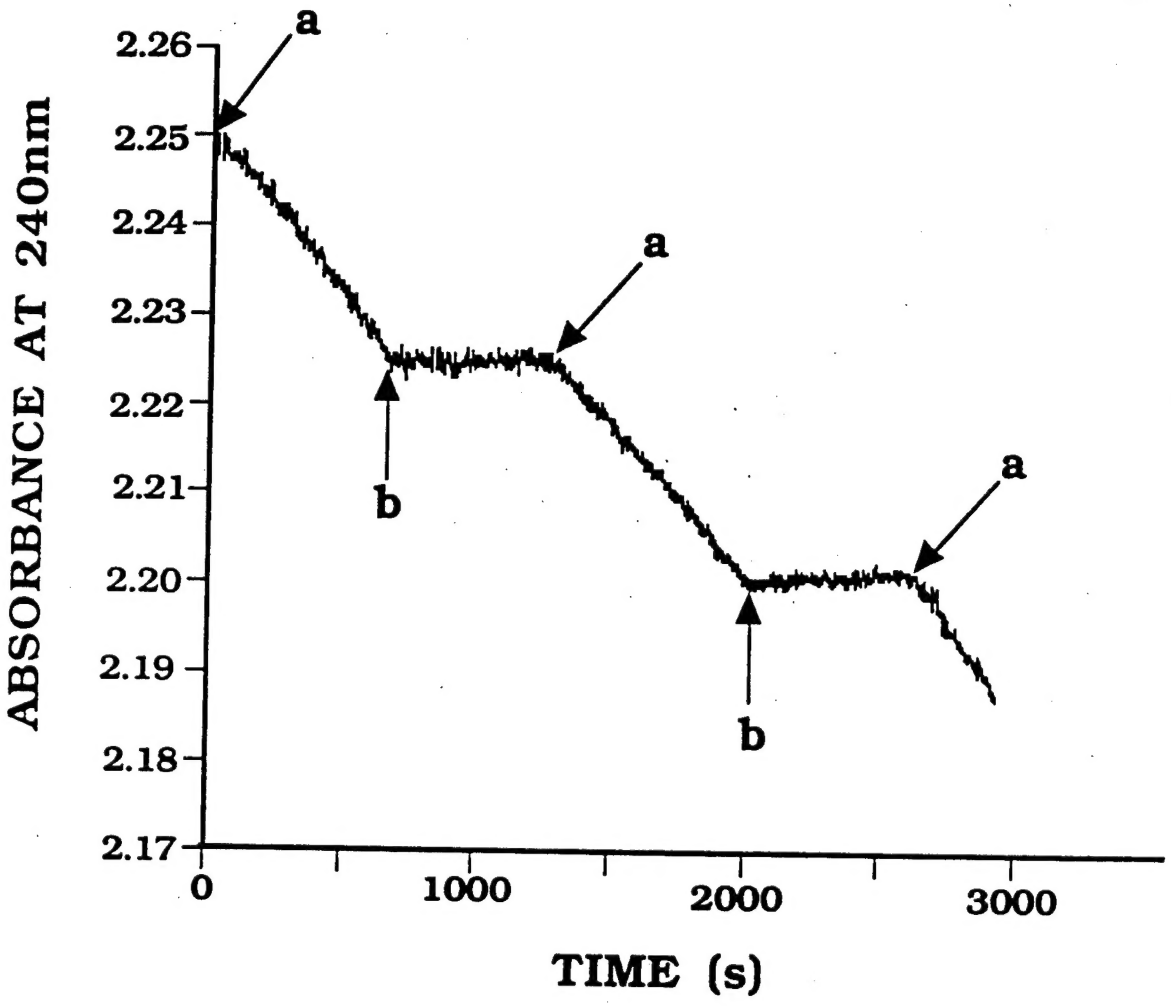

ABSORBANCE AT 240nm
2.26
2.25
2.24
2.23
2.22
2.21
2.20
2.19
2.18
2.17
0
1000
2000
3000
TIME (s)
a
a
a
b
b

Fig 13